Updates and Practical Approaches to Reproductive Disorders in Reptiles

John M. Sykes IV, DVM, DACZM

KEYWORDS

• Reptile • Reproduction • Dystocia • Chelonian
• Lizard • Snake • Infertility

Reproductive biology and disorders are important facets of captive reptile management. When presented with any type of reptile patient, the clinician should investigate the specific biology of that species to help identify the potential environmental or husbandry-related causes of reproductive disorders. Although factors and conditions for normal reproduction differ between species depending on their natural histories, the general principles and commonalities are discussed in the following sections. Reproductive disorders are relatively common in captive reptiles.[1,2] Disorders addressed in this review include infertility or lack of conception, follicular stasis, dystocia, and reproductive organ prolapse. This article is divided into sections based on the taxonomic group, although many of the predisposing factors for and the approaches to these conditions are similar for all groups.

CHELONIANS

Normal Reproduction

In males, sperm is produced seasonally, depending on the natural history of the species. During breeding season, the testicles enlarge. The relaxed phallus is located within a groove in the ventral portion of the cloaca. Neither is the cloaca connected to the urinary system nor is it involved in excretion. During erection, the phallus becomes engorged and extends ventrally and cranially; it does not evaginate like the hemipenes of lizards and snakes (see discussion in the sections on lizards and snakes). The male mounts the female from behind. Sperm is delivered to the male's cloaca via the vas deferens and is then carried along the outer groove of the erect phallus to the female's cloaca. The phallus is then retracted but does not invaginate.[3–5]

Global Health Program, Wildlife Conservation Society, 2300 Southern Boulevard, Bronx, NY 10460, USA
E-mail address: jsykes@wcs.org

Vet Clin Exot Anim 13 (2010) 349–373
doi:10.1016/j.cvex.2010.05.013 vetexotic.theclinics.com

Normal reproductive cycles and biology of female chelonians have been extensively reviewed elsewhere.[4,5] In brief, folliculogenesis generally begins with increased estrogen production, which stimulates the production of vitellogenin by the liver. Yolk is produced in the liver, transported by the blood, and deposited in the oocytes. Increased levels of testosterone, triglycerides, cholesterol, and calcium may be noted during this phase. The trigger for ovulation is poorly understood, and ovulation does not seem to require copulation in most species, even though the presence of a male may be required in some species (eg, loggerhead sea turtles [*Caretta caretta*][6] and some *Testudo* spp[4]). Ova then enter either oviduct and become fertilized. It is to be noted that females may store sperm for an extended period and produce fertile eggs long after being separated from males. Albumen and shell are then added. The time for which eggs are retained in the oviduct ranges from 9 days to 6 months, although most species retain eggs for 1 to 2 months.[3] Females typically excavate a nest, deposit and cover the eggs, and then leave.[5] All chelonians are oviparous.[4]

Reproductive Disorders

Infertility

Individuals may exhibit a lack of desire or apparent inability to mate for many reasons. Medical causes may include reproductive pathology such as phallic infection or trauma. Underlying nonreproductive pathologic conditions may also result in lack of mating, so an evaluation of the entire animal is warranted. Husbandry-related causes can include lack of appropriate environmental conditions such as hibernation or rainy periods, incompatible size difference between male and female, or dominance by other individuals in the enclosure.[3] Long-term cohabitation may also suppress mating behavior; however, mating may occur if a pair is separated and then reunited. One should ensure that the individuals are of the same species and that the gender has been correctly determined.

Mature chelonians are often sexually dimorphic. Males typically have a longer tail with a more distal cloacal opening (**Fig. 1**) and a concave plastron (**Fig. 2**).[3–5] Freshwater (emydid) male turtles often have long nails on the front feet.[4,5] Some individual species have gender-specific traits such as red irises of male eastern box turtles (*Terrapene carolina carolina*) (**Fig. 3**) or long nails on the hindlimbs of female leopard tortoises (*Geochelone pardalis*). For species that have little dimorphism or for immature animals, other techniques are required to identify the gender. Males may be identified by elevated levels of plasma testosterone,[7,8] although females may have elevated testosterone levels during some stages of the reproductive cycle.[5] Karyotyping or genetic blood sexing of juveniles, as performed in many species of birds, is often not possible in chelonians because many species have temperature-dependent sex determination,[9] although some temperature-dependent species do have genetic differences between the sexes.[10] Ultrasonography may be used to identify gonads,[4,11] although this technique may be impractical with small individuals, and differentiation between quiescent ovaries and testicles may be challenging. Scanning with the animal immersed in a water bath can improve image quality, particularly in smaller individuals.[5] Laparoscopy has been used to reliably sex juvenile or hatchling turtles[7,12–14] and may become the preferred technique because it provides immediate results, can be performed quickly in small individuals without the need of a laboratory or blood collection, and offers the opportunity for biopsy in inconclusive cases. Techniques without the use of insufflation have been described,[7,15] although the use of lactated Ringer's solution for insufflation may provide better visualization and does not inhibit buoyancy in aquatic animals after the procedure.[14] Juvenile ovaries are located in the caudal coelom just cranial to the kidneys. They are thin and elongate, and the follicles

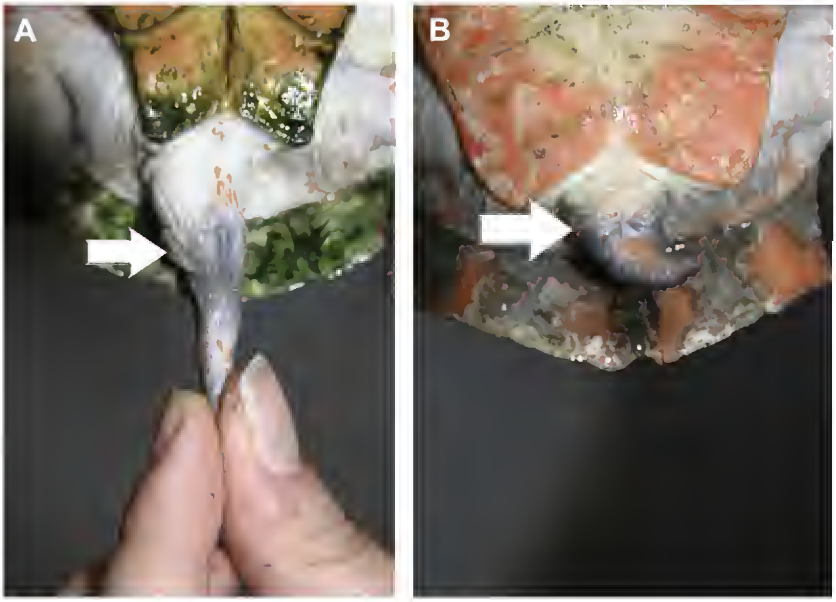

Fig. 1. Ventral views of the cloaca and tail of a male (*A*) and female (*B*) red short-neck turtle (*Emydura subglobosa*). Note the longer tail and more distal cloacal opening (*arrows*) of the male compared with the female.

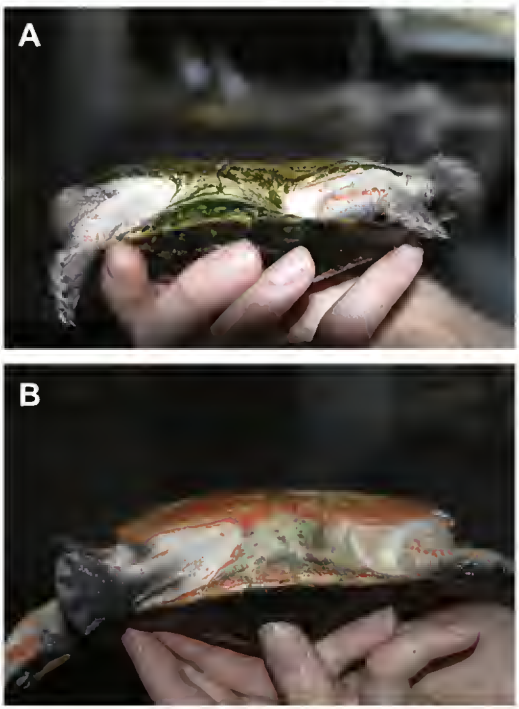

Fig. 2. Lateral views of a male (*A*) and female (*B*) red short-necked turtle (*E subglobosa*) held in dorsal recumbency. Note the concave plastron of the male and the convex plastron of the female.

Fig. 3. Iris of a male eastern box turtle (*T carolina* subsp *carolina*). Males have red or bright orange iris, whereas females have brown or light-colored iris.

may be seen even in very young individuals.[14] Testicles are pink or yellow, oval, and located cranial to and extend ventral to the kidneys.[3] A study of endoscopic examinations found pain scores to be higher in animals receiving only local anesthetics when compared with those receiving general anesthesia; therefore, these procedures should be performed only under general anesthesia.[14]

If copulation has occurred but viable eggs are not produced, spermatogenesis may be investigated. Sperm may be collected by swabbing the cloaca of the male or female after copulation or found in reproductively active males' urine.[3] Electroejaculation has been performed in multiple species.[16,17] A report of the technique described anesthesia with propofol followed by successful semen collection in leopard tortoises.[18] Alternatively, specimens may be collected for testicular biopsy using laparoscopy. Males may produce sperm only seasonally, so multiple biopsies are required to confirm lack of spermatogenesis.[3] Although some work has been done,[13,17] semen characteristics other than the presence or absence of sperm have yet to be fully characterized in chelonians. As mentioned earlier, causes for aspermatogenesis may include lack of appropriate environment or underlying medical or nutritional disorders.

Females may not produce eggs after an apparent normal copulation with sperm-producing males. Lack of environmental triggers may be the cause. Alternatively, poor body condition or nutritional disease may inhibit folliculogenesis. Ultrasonography may be used to determine if folliculogenesis is occurring and to monitor the reproductive cycle.[3,6,19] If eggs are produced but fail to develop, uterine pathology is possible, but improper incubation parameters are more likely. It has been proposed that repeated radiography of free-ranging chelonians during sensitive stages of gamete and embryo development may cause damage to the germlines and/or embryos, increasing the risk of decreased fecundity or long-term genetic problems.[20] Although there are little data evaluating this risk, whenever possible it may be prudent to use ultrasonography rather than radiography for routine reproductive assessments. However, when evaluating individuals for reproductive disorders, radiography provides information critical to decision making (see later sections) and should be used in those situations.

Eggs that fail to develop may be fertile with inappropriate incubation or infertile. Differentiating between the 2 diagnoses can be difficult or impossible. Parameters that may influence incubation include temperature, humidity, substrate, ambient oxygen, and carbon dioxide levels.[3,21] Some species may require changes in incubation temperatures, such as a cooling period, before embryonic development.[22] Eggs that do not develop should receive a necropsy, including cultures.

Follicular stasis

Follicular stasis (also called preovulatory egg-binding or retained follicles) is commonly reported in lizards but can also occur in chelonians.[3,23] Possible causes include inappropriate nutrition or environmental conditions. Recent exposure to a male after prior isolation may lead to stasis.[24] Follicles that neither ovulate nor regress can become inspissated or necrotic and may progress to yolk coelomitis (**Fig. 4**). Clinical signs can include anorexia or lethargy,[24] and clinical pathologic findings may include elevated levels of calcium, albumin, total protein, and alkaline phosphatase, with anemia, leukopenia, and heteropenia.[23] Diagnosis can be made using ultrasonography to demonstrate the persistent presence of nonovulated follicles retained in the ovary. The recommended treatment at present is ovariectomy.[3] Recovery after ovariectomy in chelonians may be prolonged when compared with an iguana with the same condition.[24] A recent technique for celioscopy-assisted ovariectomy has been described.[25] Using this method, an endoscope is placed into the coelomic cavity of the anesthetized animal through an incision in the prefemoral fossa. The ovary is grasped and gently pulled out of the prefemoral incision; oocentesis or enlarging the prefemoral incision may need to be performed to exteriorize larger follicles. The ovarian vessels are then ligated, and the ovary is removed. Ovaries of immature animals may be too tightly attached to allow for exteriorization and would require intracorporeal surgery to remove, if a prophylactic ovariectomy was desired in a young animal.[25]

Dystocia

Dystocia has been defined as a failure to lay eggs within the period that is normal for a particular species and is often referred to as egg retention.[3,26] Because a normal

Fig. 4. Gross necropsy of a cape tortoise (*Homopus* sp) that died of yolk coelmoitis. Note the free yolk in the coelomic cavity and the associated adhesions. (*Courtesy of* Dr Ed Ramsay, DVM, Knoxville, TN, USA.)

gravid female can retain eggs in the uterus for an extended period, distinguishing between normally and pathologically retained eggs can be difficult.[26] Eggs may remain within the uterus well beyond the time they should normally be deposited, and cause no overt pathologic conditions. However, sequelae of chronically retained eggs can include debilitation, infectious salpingitis, rupture of the oviduct with resulting coelomitis (see **Fig. 4**), or urinary or colonic obstruction because of oversized eggs.

Husbandry parameters (such as failure to provide an appropriate nesting site, substrate, temperature, and humidity), in addition to social factors (such as competition for nest sites or interindividual aggression), may lead to egg retention.[27] Recommendations for the appropriate depth of substrate for nesting sites vary, but depths of at least 1 to 2 times the length of the carapace have been recommended.[3] Loosening the soil or the substrate may help the animal in digging the nest.[3] Medical causes for dystocia include mechanical obstruction, reproductive tract infections, nutritional deficiencies, or an underlying systemic illness.[3,27]

In the early stages of egg retention, often, no clinical signs other than being past the due date are noted. Some individuals may pass 1 or 2 eggs but not an entire clutch.[3] These cases can be treated conservatively or may not require treatment at all. As the eggs are retained for longer periods, other signs may develop either due to the retention of the eggs or due to a predisposing condition, including anorexia, lethargy, straining, cloacal discharge, constipation, urinary obstruction, cloacal prolapse, or hind-end paresis.[3,26]

Diagnosis of dystocia can be made based on the knowledge of a particular species' normal egg-retention time or an owner's previous experiences with a particular species. Eggs may be palpated in the inguinal area but should be distinguished from cystic calculi. Radiography is useful to identify the number, position, and apparent shell quality of eggs,[3] in addition to identifying possible predisposing factors such as metabolic bone disease or pelvic fractures/stenosis. Overly large or misshapen eggs may not be laid (**Fig. 5**), and very thick shells may indicate prolonged retention.[3] Ectopic eggs located in the coelom or bladder may be identified via radiography or ultrasonography,[28] although this may be a difficult distinction to make before surgery. Evaluation of underlying conditions should be conducted via routine blood evaluation. Hypocalcemia (ionized calcium <1 mmol/L) is often present and may be an indication of preexisting nutritional deficiencies or due to chronic calcium sequestration in the eggs. Aspiration of any coelomic effusion may be supportive of yolk coelomitis.

Chelonian dystocia is rarely an emergency and can often be resolved with husbandry changes or medical therapies. In apparently otherwise healthy individuals with a normal radiographic appearance of eggs, provision of an appropriate nest site and removal of social stressors can result in laying (oviposition). Oviposition can be medically induced using oxytocin, β-blockers, fluid therapy, and calcium supplementation. Any patient with dehydration or electrolyte imbalance should first be rehydrated and stabilized. Soaking may cure mild dehydration. For more severe dehydration, intracoelomic, intraosseous, or intravenous administration of fluids should be done before additional therapies. Medical induction of oviposition in a dehydrated or unstable animal is unlikely to be successful and may increase the risk of worsening the underlying metabolic disorders or rupturing a dehydrated friable oviduct. If the animal is hypocalcemic, parenteral calcium supplementation (intracoelomic, intramuscular, or subcutaneous administration of calcium gluconate, 50–100 mg/kg)[3,26] can aid in oviductal contractions.[26] Oxytocin, 1 to 20 IU/kg, can be administered intramuscularly[3,29–31] or, continuously, via an intraosseous catheter; the lower end of the dose range is often effective in chelonians. Oxytocin can be readministered if

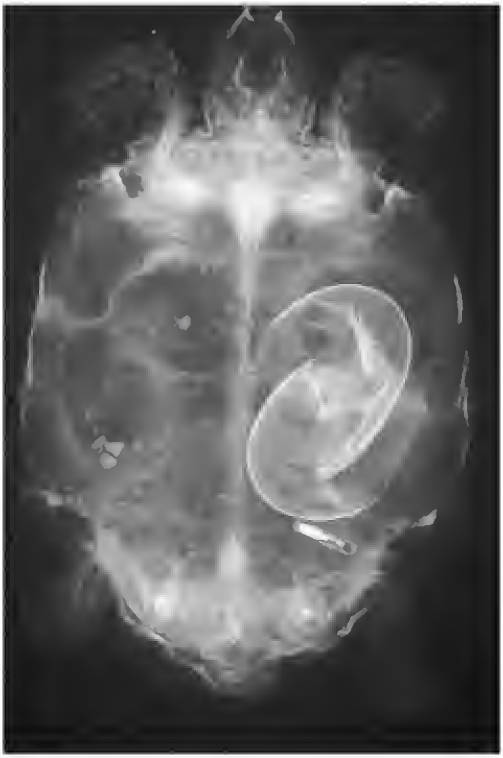

Fig. 5. Radiograph of a cape tortoise (*Homopus* sp) with a misshapen, collapsed egg within the oviduct. Eggs of this appearance should be surgically removed by either a prefemoral or a plastronotomy approach. (*Courtesy of* Dr Ed Ramsay, DVM, Knoxville, TN, USA.)

oviposition does not occur; various recommendations include administering thrice with an interval of 90 minutes with increasing doses[3] or 50% to 100% of the original dose 1 to 12 hours later.[26,32] McArthur[4] suggests the following protocol: rehydrate the patient; lubricate the cloaca; provide suitable nesting area, heat, and humidity; administer calcium, if needed, in the evening followed by atenolol (7 mg/kg by mouth) and oxytocin (1–3 IU/kg intramuscularly) the following morning; continue this protocol if eggs are being produced daily; and discontinue when oviposition stops. Arginine vasotocin is reported to be more effective in reptiles than oxytocin.[32] However, oxytocin works reasonably well in chelonians when compared with other groups of reptiles,[31] and arginine vasotocin is at present available only as a research drug.

Other adjunct medications may be beneficial, but their use has been studied little in chelonia. Prostaglandins may aid in oviposition and have been used in other reptiles.[33] A combination of oxytocin, 7.5 U/kg, and prostaglandin $F_{2\alpha}$, 1.5 mg/kg, given subcutaneously has been effective in inducing oviposition in red-eared sliders (*Trachemys scripta elegans*) (Mark Feldman, MD, personal communication, 2009), although it may be less effective in turtles that weigh more than 5 kg. Application of prostaglandin E gel on the cloaca has been recommended by Innis,[34] with no adverse or beneficial effects being noted. β-Blockers, such as atenolol (7 mg/kg) administered orally,[26] have been reported to potentiate the effects of oxytocin in chelonia. Propranolol, 1 mg/kg, intracoelomically has been used in lizards and may also be beneficial in chelonians.[33] Partial oviposition in 2 snapping turtles (*Chelydra serpentina*) sedated with

medetomidine, 150 μg/kg, for repair of traumatic injuries has been reported.[34] Use of medetomidine may aid in relaxation of sympathetic tone in stressed individuals. Eggs that have progressed to the pelvic canal but have not been deposited may be expulsed via digital palpation of the patient within the prefemoral fossa under sedation.

More aggressive therapies are warranted, when conservative management is unsuccessful, abnormally shaped or sized eggs are present (see **Fig. 5**), and the animal shows signs of debilitation or is straining. Salpingotomy may be performed via a plastronotomy or a prefemoral approach. A plastronotomy is required if a large field of vision is required (eg, if yolk coelomitis is present), but the prefemoral approach is less invasive. Details of plastronotomy have been reported elsewhere.[35,36] A prefemoral celiotomy for ovariectomy or salpingotomy may be performed in species with a large prefemoral area and small plastron, such as sea turtles.[37] A celioscopy-assisted prefemoral approach for ovariectomy has been described for species with smaller prefemoral openings (see earlier discussion).[25] This technique may also provide adequate access for salpingotomy or salpingectomy. For eggs retained in the urinary bladder, a prefemoral celiotomy for cystotomy may be successful.[28]

Alternatively, ovocentesis may be performed via the cloaca.[3,26] A speculum (laryngoscope or rodent oral speculum) is helpful to visualize the egg, and a large-gauge needle is used to puncture the egg and aspirate its contents.[3,26] The egg usually fractures after aspiration, and the egg fragments may pass on their own or can be removed using forceps. For punctured eggs that do not pass, a novel technique has been described. The tip of a Foley catheter is cut such that the balloon is at the end of the catheter, the amount of air needed to inflate the balloon to the appropriate size for the egg to be removed is determined, and the infusion port is filled with water and placed in the freezer to improve the rigidity of the catheter. The catheter is then placed into the egg via the centesis hole, the balloon is inflated, and traction is applied to remove the egg. Care must be taken not to overinflate the balloon or tear the oviduct during this process.[34] McArthur[26] recommends irrigating the cloaca and oviduct after this procedure and continuing oxytocin and β-blocker administration. If the egg is adhered to the uterus, salpingotomy may be required. A similar technique of cystoscopy performed via the cloaca followed by implosion of the egg and removal of the fragments has been described to remove retained eggs within the bladder.[38]

Yolk coelomitis

Yolk coelomitis can be secondary to retained follicles, oophoritis, salpingitis, or dystocia. Clinical signs are often nonspecific and include anorexia, lethargy, diarrhea, or lack of fecal/urine production. Clinical pathologic changes may include hypercalcemia, hyperproteinemia, anemia, and/or azotemia. Ultrasonography and radiography may aid in identifying predisposing factors such as follicular stasis or dystocia (**Fig. 6**). In addition, ultrasonography may aid in guiding a needle for coelomic centesis. Laparoscopy can differentiate normal smooth, uniform, spherical follicles from degenerating hyperemic, brown or purple follicles and may help visualize free fluid or adhesions. In contrast to a chelonian with retained eggs in the oviduct, diagnosis needs to be made rapidly because these patients may decline very quickly once coelomitis has begun. Treatment consists of stabilizing the patient and then performing exploratory celiotomy (**Fig. 7**). The source of the egg material needs to be removed, so an ovariectomy, salpingotomy, and/or salpingectomy may need to be performed. Histopathologic and microbiologic examinations may help elucidate a cause for the condition. The coelom should then be thoroughly flushed to remove any remaining yolk material. Supportive care and appropriate analgesic and antimicrobial therapy should be continued postoperatively.

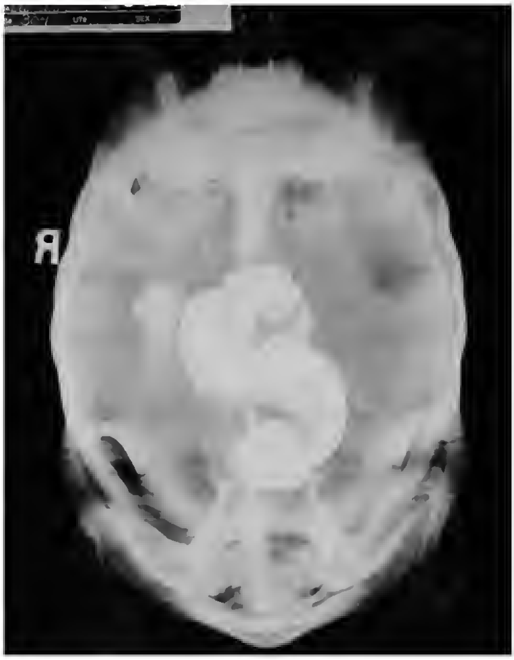

Fig. 6. Radiograph of a Burmese star tortoise (*Geochelone platynota*) with egg yolk coelomitis. The retained eggs were present unchanged within the uterus for 6 months before the sudden onset of profound lethargy and elevated uric acid level (170 mg/dL).

Phallus prolapse

Phallus prolapse may occur due to a variety of causes in chelonians (**Fig. 8**). General debilitation, neurologic dysfunction, excessive libido, trauma, lower urogenital or gastrointestinal tract infections, or causes of straining, such as constipation, parasites, gastrointestinal foreign bodies, or cystic calculi, may all predispose an animal to prolapse.[27] Nutritional metabolic bone disease may also lead to prolapse as the animal's soft tissue outgrows the shell and pushes the cloaca and phallus out.[3] A normal chelonian may erect the phallus during manipulation of the cloaca or handling of the caudal shell, but the penis should be retracted within a few hours.[3]

The prolapsed phallus should be cleaned and returned to the cloaca, if viable. Use of hypertonic solutions or cold compresses may aid in reducing swelling, and may require sedation or anesthesia. Trauma to the phallus from cagemates is common and may need to be treated before replacement of the phallus in the cloaca. Once replaced, a purse-string suture around the cloaca should be placed for 2 to 3 weeks, loose enough so that the animal can defecate but not reprolapse. If the tissue is nonviable or unable to be reduced, it may be amputated. Because the urinary system is separate from the phallus, amputation affects only the breeding potential. In small individuals, the base of the phallus can be ligated with encircling sutures or vertical mattress absorbable sutures, and the phallus is transected and removed. For larger individuals, the lateral vessels should be individually ligated and each corpus cavernosum should be separately ligated. The phallus can then be dissected free of the cloaca and transected. The cloacal tissue remaining after dissection can then be closed over the stump of the phallus in a simple continuous pattern.[3] Appropriate antibiotic and analgesic medications should be administered, and treatment of any underlying cause should be initiated.[39]

Fig. 7. Plastronotomy for egg removal, ovariosalpingectomy, and coelomic lavage for a Burmese star tortoise (*G platynota*) that was shown in Fig. 6. The animal made a complete recovery.

LIZARDS

Normal Reproduction in Lizards

Male lizards have paired hemipenes invaginated within the base of the tail. During copulation, only 1 of the 2 hemipenes is everted into the female's cloaca. Sperm is delivered to the male's cloaca via vas deferens and travels along a groove on the outside of the everted hemipenis to the female's cloaca. Similar to chelonians and snakes, the copulatory organ in lizards is not involved in excretion. Sperms within the female's reproductive tract may then fertilize ova or, in some species, may be stored for future use. Most male lizards have seasonal testicular enlargement and breeding behaviors.[40]

Female lizards can be oviparous (eg, monitors, most iguanids) or viviparous (eg, many skinks and chameleons).[40,41] The viviparous species provide varying degrees of nutrients to the developing embryo, directly via the placenta or indirectly via the preovulatory yolk.[41] Some species can be parthenogenic.[40]

Reproductive Disorders

Infertility

Causes of infertility may be similar to those for chelonians, although there is little published material addressing this concern in lizards. Correct identification of gender is important. Some species are dimorphic and gender can be readily identified but

Fig. 8. Prolapsed phallus of an unknown species of tortoise. If unable to replace, the chelonian phallus can be surgically removed from its base without affecting urination. (*Courtesy of* Dr Jean Paré, DMV, DVSc, New York, NY, USA).

many are not. Some males can be identified by a hemipenal bulge at the base of the tail.[40,41] The vent can be probed in a similar manner as described in snakes in the following sections.[40] Femoral pores of male iguanids are generally larger than those of the females (**Fig. 9**).[41] Many species of monitor lizards (*Varanus* spp) have mineralization of the hemipenes, sometimes with an internal skeleton called hemibaculum, which can be identified radiographically (**Figs. 10** and **11**).[40,41] Follicles and possibly hemipenes may be identified using ultrasonography, and celioscopy can be used to confirm the gender.[15,42]

Fig. 9. Ventral thighs of a male desert iguana (*Dipsosaurus dorsalis*). Note the line of enlarged femoral pores that identify this individual as a male (*arrows*).

Fig. 10. Ventrodorsal radiograph of the tail and pelvic region of a quince monitor lizard (*Varanus melinus*). The mineralized hemibaccula in this individual (*arrows*) identifies it as a male.

Sperm evaluation and assisted reproductive techniques have not been extensively studied in lizards, although some work describing electroejaculation has been performed.[43] Hemipenal casts or plugs may interfere with normal copulation. These casts are formed from shed skin, sperm, and exudates (**Fig. 12**),[44] and can be gently removed before the onset of breeding season.

Fig. 11. Lateral radiograph of the tail and pelvic region of a quince monitor lizard (*V melinus*). The mineralized hemibaccula in this individual (*arrow*) identifies it as a male.

Fig. 12. Hemipenal casts of a bearded dragon (*Pogona vitticeps*). These casts are formed from glandular secretions and shed skin, and occur in lizards and snakes. The casts should be removed at the start of the breeding season to aid in successful copulation. (*Courtesy of* Dr Jean Paré, DMV, DVSc, New York, NY, USA.)

Follicular stasis (preovulatory egg binding)

Similar to chelonians, inappropriate husbandry conditions, including lack of appropriate environmental cues, lack of nesting substrate, and poor nutrition, can lead to follicular stasis or dystocia (postovulatory egg binding). In follicular stasis, follicles are produced by the ovary but fail to ovulate. Lack of an adequate nesting site seems to be a common cause of egg retention in iguanas (*Iguana iguana*),[32] in which this condition is particularly common.[1,45] Some lizards may normally have a long preovulatory follicular development,[46] which needs to be distinguished from stasis. Clinical signs of stasis include anorexia and lethargy; abdominal distention may also be observed.[32] Normal gravid iguanas may have decreased food consumption, particularly while nearing ovulation, and may lose some body condition during this period,[46] but persistently anorexic or emaciated animals are likely to be in stasis. Although follicles may be resorbed in some individuals, if left untreated, retained follicles may rupture, leading to yolk coelomitis, profound debilitation, and death. Gravid lizards that are depressed or unresponsive are likely to be in stasis or in dystocia, and should be rapidly evaluated and supported. In a captive collection of Fiji Island banded iguanas (*Brachylophus fasciatus*), coelomitis caused by yolk that leaked from preovulatory follicles undergoing atresia was a leading cause of death in sexually mature females.[47] In that collection, clinical signs ranged from none to skin discoloration and coelomic distention, although several animals had had previous reproductive disorders (eg, dystocia, lethargy associated with reproductive activity, and small or no clutches in the previous years).[47] Yolk coelomitis has also been identified as a major cause of death in captive Komodo dragons (*Varanus komodoensis*).[48] Specific causes for lack of ovulation or normal atresia without resulting in coelomitis have yet to be determined, but some type of behavioral or environmental cue is thought to be involved.

Diagnosis may be made using radiography or ultrasonography to confirm the presence of follicles. A space-occupying mass may be seen with radiographs in the mid-coelom to caudal coelom. A pneumocoelogram (created by injection of air into the coelom) may be used if follicles cannot be visualized on a plain radiograph,[40] although ultrasonography may be a faster and less-invasive methodology. Blood should be drawn to assess for underlying or concurrent conditions. Clinical pathologic

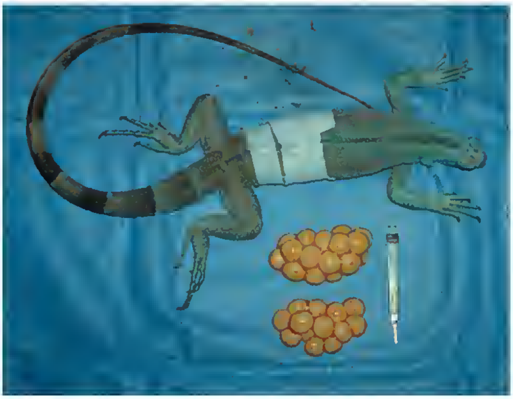

Fig. 13. Green iguana (*I iguana*) after ovariectomy for follicular stasis. Note that these retained follicles can occupy much of the available coelomic space. (*Courtesy of* Dr Ed Ramsay, DVM, Knoxville, TN, USA.)

abnormalities may include elevated levels of serum calcium and phosphorus[49] or indications of dehydration (hemoconcentration), although lizards with normal vitellogenesis may also have elevated levels of blood calcium and/or phosphorus[1] and some affected animals have normal clinical pathologic findings.[50] Heterophilia, particularly if there are toxic changes, may indicate marked inflammation and/or an infectious component.[1]

The treatment for follicular stasis is ovariectomy (**Fig. 13**). Induction of ovulation in a lizard (*Anolis carolinensis*) in a laboratory setting using follicle-stimulating hormone has been reported,[51] but to the author's knowledge, medical induction of ovulation in sick lizards with follicular stasis has not been described. The patient should be stabilized, which may require intravenous or intraosseous fluid administration. A ventral midline incision for celiotomy can be made, even though some investigators prefer a paramedian incision to avoid the ventral vein, which is suspended cranially just deep to the skin along the midline from the umbilicus (**Fig. 14**).[40,52] The ovary can

Fig. 14. Ventral abdominal vein of an unknown species of lizard. Note the close apposition of this vein with the ventral midline of the lizard. Many surgeons prefer to make a paramedian incision for celiotomies in lizards to avoid traumatizing this vein. (*Courtesy of* Dr Jean Paré, DMV, DVSc, New York, NY, USA.)

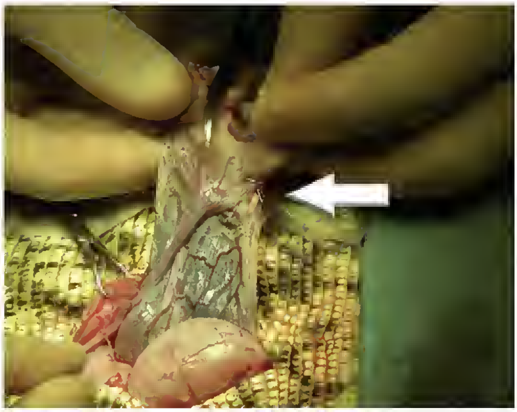

Fig. 15. Ovariectomy in a white-throated monitor lizard (*Varanus albigularis*) because of lethargy and oophritis. Note the use of sequentially placed hemoclips to ligate ovarian vessels and transect the pedicle (*arrow*).

be carefully retracted or exteriorized. Ovarian vessels are identified and ligated. The author prefers hemoclips to sutures for ligation because their use can significantly shorten the procedure time (**Fig. 15**). Often, both ovaries are affected and should be removed. Care should be taken to avoid damaging the vena cava when removing the right ovary and to leave the adrenal gland in place when removing the left ovary (**Fig. 16**). The oviduct can then be removed, if desired, although it may be preferable to leave the oviduct in place to shorten anesthetic time. Body wall and skin can be closed routinely. Postoperative supportive care, antimicrobials, and analgesia should be given as appropriate.

Dystocia

In this condition, follicles are ovulated into the oviduct but eggs fail to be laid. Predisposing factors can include environmental (eg, lack of appropriate nesting sites, inadequate temperature, humidity, or light cycle) or mechanical (eg, misshapen eggs,

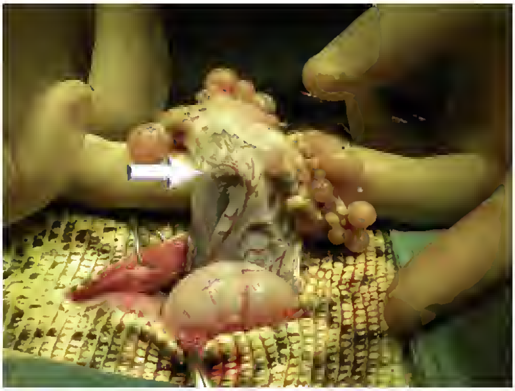

Fig. 16. Ovariectomy in a white-throated monitor lizard (*V albigularis*) because of lethargy and oophritis. Note the adrenal gland within the left ovarian pedicle (*arrow*). Care should be taken to leave this gland undisturbed.

maternal renomegaly or pelvic stenosis secondary to fractures, or metabolic bone disease) causes.[46] Affected lizards may be anorexic or lethargic, although some anorexia during gravidity is normal in many species during folliculogenesis.[46] Prolonged dystocia may result in signs consistent with hypocalcemia, such as weakness or tremors, because calcium is mobilized to the eggs (**Fig. 17**). Clinical pathologic findings may be nonspecific or may identify other predisposing factors (such as dehydration, renal disease). In a study of chameleons (*Chamaeleo chamaeleon*) with dystocia, elevation of the monocyte count and higher levels of aspartate aminotransferase (AST) were noted when compared with healthy gravid individuals.[53] The elevated level of AST was thought to be caused by tissue trauma.

Mechanical causes of dystocia can be diagnosed via cloacal examination and radiography. In animals without mechanical obstruction, medical therapy may be attempted. Oxytocin (5–30 IU/kg[32]) can be used, although it is less effective in lizards compared with chelonians. Arginine vasotocin, as the reptile equivalent of oxytocin,

Fig. 17. Radiograph of a gravid leaf-tail gecko (*Uroplatus henkeli*). Note the thick shell on the eggs. This animal displayed signs of hypocalcemia (tremors, weakness), and a salpingotomy was performed to remove both eggs.

can be more effective but is available at present only as a research drug. In healthy, gravid, striped plateau lizards (*Sceloporus virgatus*), oviposition was induced using propranolol (1 μg/g) alone or followed by arginine vasotocin (500 ng/g) or prostaglandin $F_{2\alpha}$ (25 ng/g).[33] However, propranolol and prostaglandin $F_{2\alpha}$ were less successful in green iguanas.[54] Future investigation into the use of β-blockers and prostaglandins in lizards may be informative.

Similar to preovulatory follicles, eggs within the oviduct may rupture, leading to yolk peritonitis.[49] If medical therapies are unsuccessful in relieving the dystocia or if there are mechanical obstructions, salpingotomy is indicated. The approach to the oviduct is similar to that described for ovariectomy discussed earlier. The eggs may be removed from the oviduct or oviducts, and the oviduct is closed using absorbable suture in a simple continuous pattern.[52] If eggs are adhered to the oviducts or if they are damaged or ruptured, an ovariosalpingotomy should be performed. If the oviduct is removed, the ipsilateral ovary should also be removed to prevent ovulation into the coelomic cavity and subsequent coelomitis.

Hemipenal prolapse

Prolapse of the hemipenes may result from infection, trauma, or excessive or aggressive breeding (**Fig. 18**). It may be possible to clean and replace the hemipenes as described previously for penile prolapse in chelonia. Often however, if the tissue is traumatized or desiccated, it should be amputated. For amputation, the prolapsed hemipenes should be fully everted and clamped at its base. Horizontal mattress sutures of an absorbable material are placed at the base, and the hemipenes are transected and removed. The stump can then be replaced into the base of the tail. In case of infection, the caudal tail area should be carefully examined or explored to ensure that all affected tissues are recognized and treated. Amputation of one hemipenis preserves the animal's ability to breed.[39,40]

SNAKES

Normal Reproduction

Male snakes have hemipenes similar to lizards with each hemipenis invaginated caudal to the cloaca. One hemipenis is used at a time during copulation. Sperm is delivered in a manner similar to that described earlier for lizards. The epididymis is reportedly absent in snakes.[41]

Fig. 18. Prolapsed hemipenes of an unknown species of gecko. If unable to replace, lizard hemipenes can be surgically removed from the base without affecting urination. (*Courtesy of* Dr Jean Paré, DMV, DVSc, New York, NY, USA.)

Female snakes can be oviparous (eg, pythons and most colubrids) or viviparous (eg, boas, all rattlesnakes, and most vipers).[41,44] Distinctions between viviparous and ovoviviparous species are no longer used, with all live-bearing snakes now being referred to as viviparous. Like lizards, some species may also be parthenogenic.[41]

Normal conditions to stimulate breeding behavior vary by the species. Similar to other groups of reptiles, temperature, humidity, light cycle, or social changes (ie, introduction of a male) may induce folliculogenesis and/or ovulation.[44] Many snakes may not reproduce if they do not have enough fat stores to produce follicles,[41] so a thorough examination before the onset of breeding season aids in successful reproduction.[44]

Reproductive Disorders

Infertility

Causes of infertility may be similar to those for chelonians and lizards, although there is little published material addressing this concern in snakes. As with other reptiles, accurate gender identification is critical for successful breeding and avoiding potential intraspecies aggression.[44] Snakes are rarely sexually dimorphic. Some exceptions include male boids, which often have larger cloacal spurs than females (**Fig. 19**), and some vipers, which may have color differences between the sexes (**Fig. 20**). In young snakes, it may be possible to evert the hemipenes by applying pressure and rolling the hemipenes out of the cloaca.[41,44] Alternatively, probing the hemipenes is a reliable method in most snakes. A round-ended sexing probe (snake sexing kits are commercially available) or a small, straight ball-tipped feeding tube can be lubricated, placed into 1 of the 2 openings at the lateral distal aspect of the cloaca, and directed caudally. Plastic-tipped straightened bobby pins have been recommended by Denardo,[41] because they can be discarded after a single use. The probe is easily

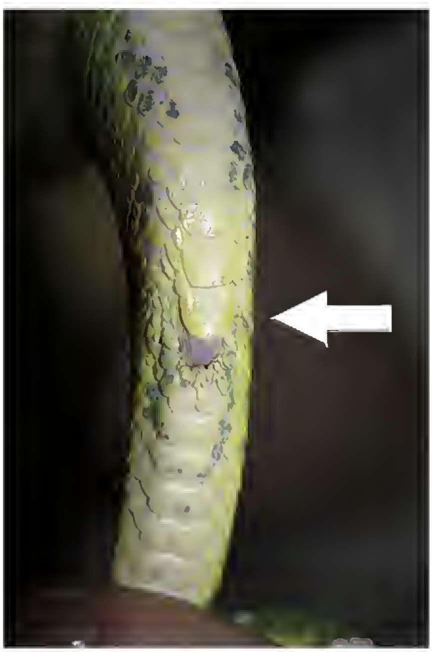

Fig. 19. Cloaca of a male emerald tree boa (*Corallus caninus*). Note the presence of cloacal spurs (*arrow*). These spurs are small or absent in females, and are a common trait in many boids.

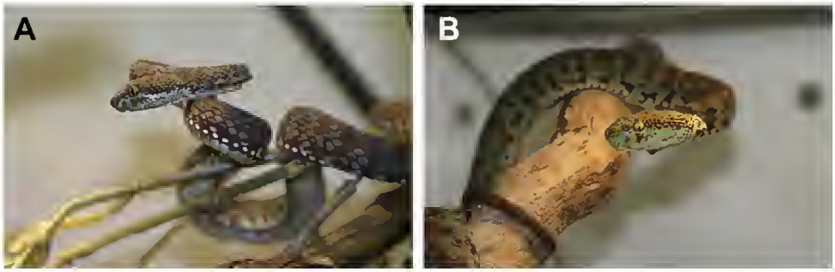

Fig. 20. Lateral views of the heads of a male (*A*) and female (*B*) Kanburian pit viper (*Trimeresurus kanburiensis*). Males have a white stripe on the lateral aspect of their head, which females lack. This stripe is present from birth.

advanced in males, much further than in females, which may have a short blind-ended sac in this area. A larger probe is preferred to a smaller one, because the larger probe is less likely to enter the female diverticula and gives an inaccurate result.[44] The number of scutes that the probe advances in males depends on the species, so it helps to have a pair of age-matched animals to compare with each other.

In animals for which manual eversion or probing is unsuccessful or indeterminate, hydrostatic eversion can be performed. A needle is inserted caudal to the end of the presumptive hemipenes, and sterile fluid is injected into the subcutaneous space. The fluid pressure causes the hemipenes and, potentially, portions of the cloaca to evert, and the hemipenes or oviductal papilla can be identified. Anesthesia may be required in larger individuals. Care should be taken to be as sterile as possible, to avoid the scent glands in this area, and to avoid injection or direct trauma to the hemipenes during this procedure.[41]

Hemipenal casts or plugs may interfere with normal copulation. These casts are formed from shed skin, sperm, and exudates (see **Fig. 12**).[44] They can be gently removed before the onset of breeding season.

Sperm assessment and assisted reproductive techniques have been studied in snakes and may become more widely used in the future.[55] Semen may be collected manually via massage[56,57] or through electroejaculation.[58]

Follicular stasis

Follicular stasis is rarely reported in snakes but may occur.[59] Treatment is ovariectomy, although multiple celiotomies may be required to remove the entire ovary (**Fig. 21**). Egg retention within the oviduct (dystocia) is much more common in snakes.

Dystocia

Causes for dystocia are similar to those described earlier for lizards. Inappropriate temperatures or humidity, lack of appropriate nest site, malnutrition, or social stressors may all contribute to dystocia. Many snakes oviposit within a covered box with damp moss or vermiculite inside.[44] For oviparous snakes, a common presentation is the recent oviposition of a clutch but retention of 1 to 2 eggs.[41,44] Retained eggs can often be palpated and possibly visualized. For viviparous species, lack of young after an expected due date may be observed. For both types of snakes, clinical signs may be absent, may be nonspecific, such as anorexia or lethargy, or may include straining or cloacal prolapse.

Retained eggs or young may be palpated or visible as swellings, but care should be taken during palpation as the oviduct can easily tear.[44] Radiographs may confirm the presence of young in viviparous species; uncurled young within the dam after the due

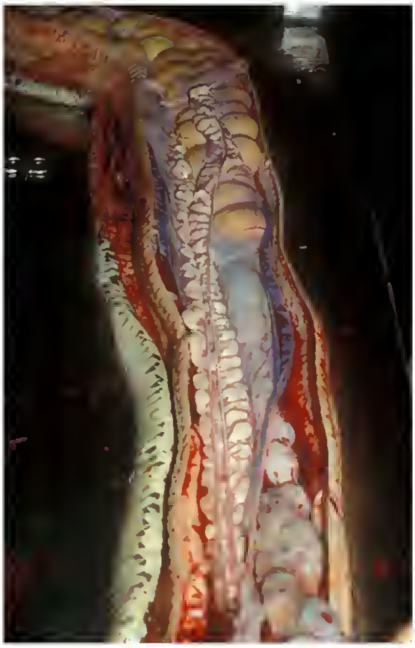

Fig. 21. Necropsy of an unknown species of python with retained follicles. Note the longitudinal extension of the ovaries. Often, multiple celiotomy incisions are required for ovariectomy in snakes because of this anatomy. (*Courtesy of* Department of Pathology, The University of Tennessee College of Veterinary Medicine, Knoxville, TN, USA; with permission.)

date or gas surrounding the young may indicate fetal death.[41,44] Plain radiographs are less useful for oviparous species as snake eggs are typically soft shelled and are poorly delineated with radiographs. However, the use of a contrast agent delivered orally or colonically may aid in identification of other structures in the caudal coelomic cavity that could be causing dystocia (**Fig. 22**). Ultrasonography may also be useful in identifying viviparous young or retained eggs, and it may be possible to detect fetal heartbeat or movement of the fetus.

Medical therapy can be attempted but is often unsuccessful. Possible obstructive causes for dystocia (eg, renomegaly or an overly large/malformed egg in the distal

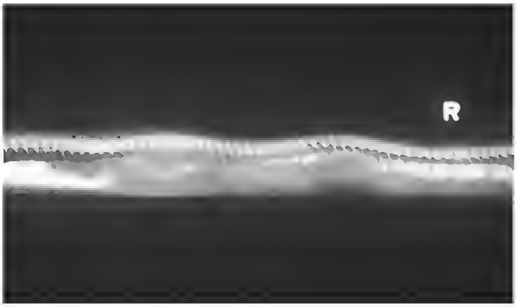

Fig. 22. Lateral radiograph of an unknown species of boa with egg masses within the oviduct. Note that the barium contrast within the intestinal tract identifies the masses as extraintestinal. (*Courtesy of* Dr Ed Ramsay, DVM, Knoxville, TN, USA.)

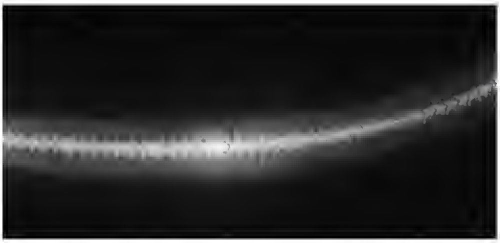

Fig. 23. Dorsoventral view of a rat snake (*Elaphe* sp) with a misshapen egg. Eggs of this shape should be surgically removed via celiotomy. (*Courtesy of* Dr Ed Ramsay, DVM, Knoxville, TN, USA.)

oviduct [**Fig. 23**]) should be first ruled out via physical examination, radiography, and possibly ultrasonography. If no obstruction is seen, oxytocin , 5 to 20 IU/kg, given intramuscularly may be effective, particularly if given within 2 to 3 days from the onset of dystocia;[44] however, in a reported case, live young were obtained after oxytocin administration 7 days after normal parturition had begun.[60] Arginine vasotocin may be more effective but is at present available only as a research drug. Use of additional medical therapies such as β-blockers or prostaglandins has received little attention in snakes.[61]

Eggs may be manually milked out of the cloaca, but this procedure carries a high risk of oviductal tearing and rupture of the follicles.[32] To be performed safely, the animal should be first anesthetized, and only gentle pressure should be applied to the eggs to move them toward the cloaca. Once near the cloaca, eggs may be visualized and aspirated/imploded via the cloaca as described earlier for chelonians. The remaining shell may be manually removed or left to pass on its own; however, care should be taken not to tear the oviduct because the shells may adhere to the oviductal mucosa if they have been in place for several days.[44] If the eggs cannot be removed medically or manually, surgery should be performed. A single paramedian celiotomy may be performed, but if there are many eggs or if they are adhered to the oviduct in multiple locations, multiple celiotomies may be necessary. Once eggs or fetuses are removed, an otherwise healthy oviduct can be closed in a simple continuous

Fig. 24. Surgery to remove prolapsed hemipenes of a rat snake (*Elaphe* sp). Note that the hemipenes have been fully everted and are ligated with 2 encircling sutures before being transected. Removal of the hemipenes does not affect excretion in lizards or snakes. (*Courtesy of* Dr Jean Paré, DMV, DVSc, New York, NY, USA.)

pattern. If excessive inflammation or previous tearing has occurred, or if there is evidence of yolk coelomitis, the oviduct and ipsilateral ovary may need to be removed. Removal of the ovary often requires a long or multiple incisions to remove the entire organ (see **Fig. 21**).[62] The coelom can be closed in a simple pattern, and the skin can be closed using everting sutures.

Prolapse

The oviduct may prolapse secondary to straining from dystocia. If eggs are still present in the prolapsed tissue, it may be possible to remove the eggs, close the oviduct, and replace it within the cloaca. However, in most cases, the oviduct is too damaged to be replaced and should be surgically removed along with the ipsilateral oviduct as described previously.

The hemipenes may prolapse because of infection, trauma, or excessive use.[44] Therapy for hemipenal prolapse is as described earlier in lizards (**Fig. 24**).

ACKNOWLEDGMENTS

The author thanks Jean Paré and Ed Ramsay for contribution of their photographs and review of the manuscript. Special thanks to Jennifer Pramuk and Megan Baumer of the Department of Herpetology, Bronx Zoo for their assistance with the photography of sexually dimorphic traits.

REFERENCES

1. Campbell TW. Clinical laboratory evaluation of dystocia in lizards. Proceedings of the Association of Reptilian and Amphibian Veterinarians 1999;123–31.
2. Lloyd ML. Reptilian dystocias review: prevention, management, and comments on the synthetic hormone vasotocin. Proceedings of the American Association of Zoo Veterinarians 1990;329–36.
3. Innis C, Boyer TH. Chelonian reproductive disorders. Vet Clin North Am Exot Anim Pract 2002;5:555–78.
4. McArthur S. Reproductive system. In: McArthur S, Wilkinson R, Meyer J, editors. Medicine and surgery of tortoises and turtles. Oxford (UK): Blackwell Publishing; 2004. p. 57–63.
5. Kutchling G. The reproductive biology of the chelonia. Berlin: Spinger-Verlag; 1999.
6. Manire CA, Byrd L, Therrien CL, et al. Mating-induced ovulation in loggerhead sea turtles, *Caretta caretta*. Zoo Biol 2008;27:213–25.
7. Rostal D, Grumbles J, Lance V, et al. Non-lethal sexing techniques for hatchling and immature desert tortoises (*Gopherus agassizii*). Herpetological Monographs 1994;8:83–7.
8. Owens DW. Hormones in the life history of sea turtles. In: Lutz P, Musick JA, editors. Biology of the sea turtles. Boca Raton (FL): CRC Press; 1996. p. 315–41.
9. Madge D. Temperature and sex determination in reptiles with reference to chelonians. BCG Testudo 1994;2(3):9–14.
10. Demas S, Duronslet M, Wachtel S, et al. Sex-specific DNA in reptiles with temperature dependant sex determination. J Exp Zool 1990;253:319–24.
11. Stetter M. Ultrasonography. In: Mader D, editor. Reptile medicine and surgery. 2nd edition. St Louis (MO): Elsevier Inc; 2006. p. 665–74.
12. Kuchling G. 2006. Endoscopic sex determination in juvenile freshwater turtles, *Trachemys scripta*, a species with temperature dependent sex determination. Biol Reprod 1999;61:1275–80 – ordered.

13. Mitchell MA, Thompson D, Burgdorf A, et al. Coelioscopy as an antemortem method for confirming temperature dependant sex determination in Blanding's turtles (*Emydoidea blandingii*). In: Proceedings of the Association of Reptilian and Amphibian Veterinarians. Milwaukee (WI): 2009. p. 136.
14. Hernandez-Divers SJ, Stahl SJ. An endoscopic method for identifying sex of hatchling Chinese box turtles and comparison of general versus local anesthesia for coelioscopy. J Am Vet Med Assoc 2009;234(6):800–4.
15. Hernandez-Divers SJ, Hernandez-Divers SM, Wilson GH, et al. A review of reptile diagnostic coelioscopy. J Herp Med Surg 2005;3:16–31.
16. Platz CC, Mengden BS, Quinn H, et al. Semen collection, evaluation and freezing in the green sea turtle, Galapagos tortoise, and red-eared pond turtle. Proceedings of the American Association of Zoo Veterinarians 1980;47–54.
17. Wood F, Platz C, Critchley K, et al. Semen collection by electroejaculation of the green turtle, *Chelonia mydas*. Brit J Herpetol 1982;6:200–2.
18. Mitchell MA, Zimmerman D, Heggem B. Collection and characterization of semen from leopard tortoises. In: Proceedings of the Association of Reptilian and Amphibian Veterinarians. Milwaukee (WI): 2009. p. 166.
19. Cheng Y, Chen T, Yu P, et al. Observations on the female reproductive cycles of captive Asian yellow pond turtles (*Mauremys mutica*) with radiography and ultrasonography. Zoo Biol 2009;28:1–9.
20. Kutching G. How to minimize risk and optimize information gain in assessing reproductive condition and fecundity of live female chelonians. Chelonian Conserv Biol 1998;3(1):118–23.
21. Innis C. Infertility and embryonic death. In: McArthur S, Wilkinson R, Meyer J, editors. Medicine and surgery of tortoises and turtles. Oxford (UK): Blackwell Publishing; 2004. p. 63–8.
22. Ewert MA. Cold torpor, diapause, delayed hatching and aestivation in reptiles and birds. In: Deeming DC, Ferguson MWJ, editors. Egg incubation: its effects on embryonic development in birds and reptiles. Cambridge University Press, New York; 1991. p. 173–91.
23. McArthur S. Follicular stasis in captive chelonian, *Testudo* spp. In: Proceedings of the Association of Reptilian and Amphibian Veterinarians. Eighth Annual Conference, Orlando (FL): 2001. p 75–86.
24. McArthur S. Follicular stasis. In: McArthur S, Wilkinson R, Meyer J, editors. Medicine and surgery of tortoises and turtles. Oxford (UK): Blackwell Publishing; 2004. p. 325–9.
25. Innis CJ, Hernandez-Divers S, Martinez-Jimenez D. Coelioscopic-assisted prefemoral oophorectomy in chelonians. J Am Vet Med Assoc 2007;230(7):1049–52.
26. McArthur S. Dystocia. In: McArthur S, Wilkinson R, Meyer J, editors. Medicine and surgery of tortoises and turtles. Oxford (UK): Blackwell Publishing; 2004. p. 316–8.
27. McArthur S. Interpretation of presenting signs. In: McArthur S, Wilkinson R, Meyer J, editors. Medicine and surgery of tortoises and turtles. Oxford (UK): Blackwell Publishing; 2004. p. 273–80.
28. Thomas HL, Willer CJ, Wosat MA, et al. Egg-retention in the urinary bladder of a Florida cooter turtle, *Pseudemys floridana floridana*. J Herp Med Surg 2001; 11(4):4–6.
29. Johnson R. Dystocia in an injured common eastern long-necked turtle (*Chelodina longicollis*). Vet Clin North Am Exot Anim Pract 2006;9:575–81.
30. Tucker JK, Thomas DL, Rose J. Oxytocin dosage in turtles. Chelonian Conserv Biol 2007;6(2):321–4.

31. Feldman ML. Some options to induce oviposition in turtles. Chelonian Conserv Biol 2007;6(2):313–20.
32. DeNardo D. Dystocias. In: Mader D, editor. Reptile medicine and surgery. 2nd edition. St Louis (MO): Elsevier Inc; 2006. p. 787–92.
33. Gross T, Guillette LJ, Gross DA, et al. Control of oviposition in reptiles and amphibians. Proceedings of the American Association of Zoo Veterinarians 1992;143–50.
34. Innis CJ. Innovative approaches to chelonian obstetrics. In: Proceedings of the Association of Reptilian and Amphibian Veterinarians, Naples, Italy: 2004. p. 1–5.
35. McArthur S, Hernandez-Divers S. Surgery. In: McArthur S, Wilkinson R, Meyer J, editors. Medicine and surgery of tortoises and turtles. Oxford (UK): Blackwell Publishing; 2004. p. 403–59.
36. Mader DR, Bennett RA, Funk RS, et al. Surgery. In: Mader D, editor. Reptile medicine and surgery. 2nd edition. St Louis (MO): Elsevier Inc; 2006. p. 581–630.
37. Nutter FB, Lee DD, Stamper MA, et al. Hemiovariosalpingectomy in a loggerhead sea turtle (*Caretta caretta*). Vet Rec 2000;146:78–80.
38. Knotek Z, Jekl V, Knotkova Z, et al. Eggs in chelonian urinary bladder: is coeliotomy necessary? Proceedings of the Association of Reptilian and Amphibian Veterinarians 2009;118–21.
39. Barten SL. Penile prolapse. In: Mader D, editor. Reptile medicine and surgery. 2nd edition. St Louis (MO): Elsevier Inc; 2006. p. 862–4.
40. Funk RS. Lizard reproductive medicine and surgery. Vet Clin North Am Exot Anim Pract 2002;5:579–613.
41. Denardo D. Reproductive biology. In: Mader D, editor. Reptile medicine and surgery. 2nd edition. St Louis (MO): Elsevier Inc; 2006. p. 376–90.
42. Roberts J, Bechert U, Huber M, et al. Determination of gender in the blue-tongued skink (*Tiliqua scincoides*) by ultrasonography and endoscopy with comparison of differentiating external morphologic parameters. Proceedings of the Association of Reptilian and Amphibian Veterinarians 2007;50.
43. Mitchell MA, Zimmerman D, Heggem B. Collection and characterization of semen from green iguanas. Proceedings of the Association of Reptilian and Amphibian Veterinarians 2009;165.
44. Stahl SJ. Veterinary management of snake reproduction. Vet Clin North Am Exot Anim Pract 2002;5:615–36.
45. Raiti P. Changing trends in diseases of the green iguana, *Iguana iguana*. Proceedings of the Association of Reptilian and Amphibian Veterinarians 1999;133–6.
46. Maxwell LK. Infectious and non-infectious diseases. In: Jacobson ER, editor. Biology, husbandry, and medicine of the green iguana. Malabar (FL): Krieger Publishing Co; 2003. p. 108–32.
47. Stacy BA, Howard L, Kinkaid J, et al. Yolk coelomitis in Fiji Island banded iguanas (*Brachylophus fasciatus*). J Zoo Wildl Med 2008;39(2):161–9.
48. Spellman LH. Medical management. In: Murphy C, Walsh T, editors. Komodo dragons: biology and conservation. Washington, DC: Smithsonian Press; 2002. p. 208–10 – ordered.
49. Backeus KA, Ramsay EC. Ovariectomy for treatment of follicular stasis in lizards. J Zoo Wildl Med 1994;25(1):111–6.
50. Scheelings TF. Pre-ovulatory follicular stasis in a yellow-spotted monitor, *Varanus panoptes panoptes*. J Herp Med Surg 2008;18(1):18–20.
51. Jones RE, Austin HB, Lopez KH, et al. Gonadotropin-induced ovulation in a reptile (*Anolis carolinensis*): histological observations. Gen Comp Endocrinol 1988;72: 312–22.

52. Lock B, Bennett RA. Anesthesia and surgery. In: Jacobson ER, editor. Biology, husbandry, and medicine of the green iguana. Malabar (FL): Krieger Publishing Co; 2003. p. 152–67.
53. Cuadrado M, Diaz-Paniagua C, Quevedo MA, et al. Hematology and clinical chemistry in dystocic and healthy post-reproductive female chameleons. J Wildl Dis 2002;38(2):395–401.
54. Innis C. Treatment with propranolol and PGF2a stimulates nesting behavior but not oviposition in a gravid green iguana. Bull Assoc Reptil Amph Vet 1996;6(2):1–4.
55. Mattson KJ, De Vries A, McGuire SM, et al. Successful artificial insemination in the corn snake, *Elaphe gutatta*, using fresh and cooled semen. Zoo Biol 2007; 26:363–9.
56. Zacariotti RL, Grego KF, Fernandes W, et al. Semen collection and evaluation in free-ranging Brazilian rattlesnakes (*Crotalus durissus terrificus*). Zoo Biol 2007; 26:155–60.
57. Fahrig BM, Mitchell MA, Eilts BE, et al. Characterization and cooled storage of semen from corn snakes (*Elaphe guttata*). J Zoo Wildl Med 2007;38:7–12.
58. Quinn H, Blasedel T, Platz CC. Successful artificial insemination in the checkered garter snake. Int Zoo Yearbook 1989;28:177–83.
59. Denardo D, Barten SL, Rosenthal KL, et al. Dystocia. J Herp Med Surg 2000; 10(2):8–17.
60. Lock BA. Treatment of interrupted parturition with delivery of viable young in a Brazilian rainbow boa, *Epicrates cenchria cenchria*. Bull Assoc Reptil Amph Vet 1998;8(2):11–3.
61. Nathan R. Treatment with ovicentesis, PGE2, and PGF2a to aid oviposition in a spotted python. Bull Assoc Reptil Amph Vet 1996;6(4):4.
62. Lock BA. Reproductive surgery in reptiles. Vet Clin North Am Exot Anim Pract 2000;3(3):733–52.